Carbon Dynamics of Surface Residue– and Root-derived Organic Matter under Simulated No-till

W. J. Gale and C. A. Cambardella*

ABSTRACT

No-till practices have the potential to increase soil organic C, but little is known about the relative contribution of surface residue and roots to soil organic C accumulation. In a simulated no-till experiment, we studied the fate of ^{14}C-labeled surface residue and in situ roots during a 1-yr incubation. Soil samples collected during the incubation were chemically dispersed and separated into five particle size and density fractions. The organic C, ^{14}C, and total N content of each fraction was determined. Alkali traps were used to measure ^{14}C losses due to respiration. After 360 d, 66% of the ^{14}C contained in the surface residue on Day 0 had been respired as $^{14}CO_2$, 11% remained in residue on the soil surface, and 16% was in the soil. In comparison, 56% of the root-derived ^{14}C in the soil was evolved as $^{14}CO_2$ and 42% remained in the soil. The large (500–2000 μm) and small (53–500 μm) particulate organic matter (POM) fractions together contained 11 to 16% of the initial root-derived ^{14}C in the soil. In contrast, POM contained only 1 to 3% of the inital surface residue–derived ^{14}C. These data show clear differences in the partitioning of surface residue– and root-derived C during decomposition and imply that the beneficial effects of no-till on soil organic C accrual are primarily due to the increased retention of root-derived C in the soil.

The soil C pool in agroecosystems is receiving increased attention because of concerns about the rising levels of atmospheric CO_2. Cropland may function as either a sink or source for CO_2 in the atmosphere (Karlen and Cambardella, 1996). If we are to manage the C balance in cropland, we must understand the factors that determine whether the C contained in crop residue is retained in the soil or released as CO_2 into the atmosphere (Buyanovsky and Wagner, 1987).

The use of conservation tillage, including no-till, is being encouraged as part of a strategy to reduce C loss from agricultural soils (Kern and Johnson, 1993). Decomposition rates are generally slower in no-till compared with conventional tillage in which the decomposition of soil organic matter (SOM) is promoted by the stirring of the soil and alterations in the soil microclimate (Parton et al., 1996). Holland and Coleman (1987) suggested that C retention is increased in no-till because the surface residue is primarily decomposed by fungi which have a higher assimilation efficiency than the bacteria which dominate the decomposition processes of residue mixed into the soil. We could find no information in the literature which compared the relative contribution of surface residue and roots to soil organic C in no-till.

We conducted an experiment to examine changes in soil C pools during the initial period following the implementation of no-till. We were interested in learning more about the labile C pools associated with biologically active processes such as nutrient cycling and soil aggregation. Particulate organic matter consists of partially decomposed plant residues and is considered to be the first intermediate pool in the decay continuum between crop residue and humified, stable organic matter (Gregorich and Janzen, 1996). Cambardella and Elliott (1993) reported a strong correlation between POM C and macroaggregates in soil. The importance of POM as a source of energy and nutrients in the soil ecosystem has been emphasized by several researchers (Gregorich and Janzen, 1996; Karlen and Cambardella, 1996; Gregorich et al., 1994), however, the dynamics of POM are not well understood.

The specific objectives of this work were: (i) to determine the relative contribution of surface residue and in situ roots to SOM immediately following the implementation of no-till and (ii) to characterize the partitioning of surface residue– and root-derived C among labile SOM fractions.

MATERIALS AND METHODS

Experimental Setup

Soil from the surface horizon (0–15 cm) of a Monona silt loam (fine-silty, mixed, mesic Typic Hapludolls) was collected from a research site near Treynor, IA, in August of 1995. The soil contains 5% sand and 27% clay. The field had been continuously cropped to corn for more than 30 yr using conventional tillage practices.

The field-moist soil was passed through an 8-mm sieve. Large pieces (>~2 cm) of plant residue were removed by hand. The soil was thoroughly mixed and the equivalent of 2420 g of dry soil was placed in 2.5-L plastic pots. A nutrient solution was added to each pot at the beginning of the experiment, and identical fertilizer additions were made 18, 30, 38, 47, and 52 d after the plants emerged. The total amount of fertilizer added to the pots was similar to common field application rates for oat (67 kg N ha^{-1}, 56 kg P ha^{-1}, 22 kg K ha^{-1}). Twelve oat (*Avena sativa* cv. Ogle) seeds were planted in each pot and after emergence the plant population was thinned to six plants per pot.

The pots were placed in two growth chambers which were set at 20 and 12.7°C during light and dark periods, respectively. The plants received 10 h of light each day for the first 5 wk of the incubation and then daylength was increased to 14 h. Fifteen pots were arranged in each chamber in a completely randomized design. Additional pots were prepared as described above and placed as a border around the experimental pots to minimize edge effects.

USDA-ARS National Soil Tilth Laboratory, 2150 Pammel Dr., Ames, IA 50011. Received 30 June 1998. *Corresponding author (cindyc@nstl.gov).

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Abbreviations: hPOM, heavy particulate organic matter; LSD, least significant difference; POM, particulate organic matter; SOM, soil organic matter.

Pulse Labeling

We pulse labeled the plants in one growth chamber with $^{14}CO_2$ at 15, 20, 26, 31, 37, 46, and 52 d after emergence. No label was applied after Day 56 because the plants had reached boot stage and most new photosynthate would be used to produce grain, rather than leaf, stem, or root biomass. To label the plants, we placed a Plexiglas[1] box (0.05 m^3) over each pot. A beaker containing 270 μBq $NaH^{14}CO_3$ (New England Nuclear Research Products, Boston, MA) was placed inside the box and the base was sealed with duct tape. The $^{14}CO_2$ was generated in the box by acidifying the $NaH^{14}CO_3$ with 9 mL of lactic acid which was added to each beaker through a small injection port in the side of the Plexiglas box. The boxes were kept in place for 5 h to allow the $^{14}CO_2$ to be assimilated by the plants. The plants in the second growth chamber were not labeled with $^{14}CO_2$.

Incubation

Treatment Combinations

We used the ^{14}C labeled and unlabeled plants to create a reciprocal transplant experiment consisting of two treatment combinations: (1) ^{14}C labeled surface residue + pots with unlabeled roots and soil and (2) unlabeled surface residue + pots with ^{14}C labeled roots and soil.

Combination 1 will be referred to as the labeled surface residue treatment and Combination 2 will be referred to as the labeled (roots + soil) treatment. Note that the labeled (roots + soil) treatment contained ^{14}C prior to the start of the incubation because the plants had been exposed to $^{14}CO_2$ during vegetative growth. Therefore, measurements from the first sample date (Day 0) served as baseline values for each of the treatments to which subsequent sample dates were compared.

At senescence (118 d after planting), the plants from both growth chambers were cut off at the soil surface. The oat grain was removed and the remaining straw was dried at 50°C for 36 h. The dry weight of the straw averaged 20.2 g pot^{-1}. Leaves were separated from the stems, composited as unlabeled and ^{14}C labeled samples, and dried at 50°C for 36 h. The roots and soil were undisturbed in the pots to simulate no-tillage. The soil in each pot was adjusted to 250 g H_2O kg^{-1} dry soil and then 4.5 g subsamples of the leaves (representing 22% of the total aboveground C) were put on the soil surface. The labeled surface residue treatment was prepared by placing ^{14}C labeled leaves on the soil surface of pots containing unlabeled roots and soil. The labeled (roots + soil) treatment was prepared by placing unlabeled leaves on the soil surface of pots containing ^{14}C-labeled roots and soil. We applied 10 g of water with an atomizer to moisten the surface residue. Each pot was placed in a 15-L plastic container (Letica Corp., Muscatine, IA) that had a removable air-tight lid. The containers were arranged in a completely randomized design in one growth chamber at 25°C. The lids were periodically removed to maintain aerobic conditions within the containers. Water was added to the pots as needed to maintain the desired soil moisture.

Soil Respiration

A wide mouth glass jar (225 mL) containing 50 mL NaOH was placed in each container to capture respired CO_2. The CO_2 traps were changed on Days 1, 3, 5, 8, 12, and then weekly or biweekly for the remainder of the incubation. The concentration of the NaOH in the traps was adjusted during the incubation so that at least 40% of the trap capacity remained unused. Respired $^{14}CO_2$ was measured by adding Optima Gold (Packard Instrument Co., Downers Grove, IL) scintillation cocktail to 0.5 mL aliquots taken from each trap and counting on a 1900 TR Liquid Scintillation Analyzer (Packard Instrument Co., Downers Grove, IL). Cumulative respired $^{14}CO_2$ is reported as an average of all the pots within the labeled surface residue or labeled (roots + soil) treatments.

Soil Sampling

Three pots from each treatment were destructively sampled on Days 0, 90, 180, 270, and 360. After the surface residue was removed, the plastic pots were cut away, and the soil was gently picked apart. The coarse roots (>2 cm length) were removed and rinsed with water, although most roots were clean when separated from the soil. The surface residue and coarse roots were dried at 50°C for 48 h and then separately ground in a ball mill. After removing the coarse roots, the moist soil from each pot was passed through an 8-mm sieve and systematically mixed to ensure representative subsampling. Three 400-g samples were removed from each pot and passed through a 2-mm sieve. These samples and the remaining 8-mm sieved soil were air dried and stored at room temperature.

Size and Density Separation

Soil Dispersion and Size Separation

The air-dried, 2-mm sieved soil from each pot was systematically mixed and three samples were removed to determine organic C, ^{14}C, and total N in the whole soil. In addition, we dispersed three 30-g soil samples from each pot with 100 mL of 5 g L^{-1} sodium hexametaphosphate and shook the samples for 18 h on a reciprocal shaker. The dispersed soil samples were passed sequentially through a 500- and 53-μm sieve and rinsed thoroughly with water. The (silt + clay) material that passed through the sieves was captured in a receiving pan and then dried at 70°C for 36 h. The dried (silt + clay) fraction and the whole soil samples were ground on a roller mill to pass through a 250-μm sieve and then stored at room temperature.

We observed that the material retained on the 53-μm sieve became stuck together when it was dried in an oven. We were concerned that this would interfere with the density separation procedure. We did not observe the same tendency for the material retained on the 500-μmm sieve. Because of these observations, the material retained on the two sieves was handled differently.

Density Separation of Large Particulate Organic Matter

The material retained on the 500-mm sieve was transferred to an aluminum weighing pan and dried at 50°C. At a later date, these samples were transferred to 100 mL beakers and suspended in sodium polytungstate (Geoliquids Inc., Prospect Heights, IL) adjusted to a density of 1.85 g cm^{-3}. The samples were allowed to separate overnight, after which we aspirated the large POM floating on the surface of the liquid. The large POM was washed with 300 mL of water on a 20-mm nylon filter, transferred to aluminum weighing pans, dried at 50°C, ground in a ball mill, and then stored at room temperature.

Density Separation of Small Particulate Organic Matter

We developed a procedure that permitted flotation of the 53-μm fraction without oven drying the material first. The material retained on the 53-μm sieve was backwashed onto a 20-μm nylon filter, and a vacuum was applied to remove

[1] Reference to trade names and companies is made for information purposes only and does not imply endorsement by the USDA.

excess water. Then the material was rinsed into 100-mL beakers with polytungstate (1.85 g cm^{-3}) to a volume of 50 mL. The samples were allowed to separate overnight, after which we aspirated the small POM floating on the surface of the liquid. The small POM was rinsed, dried, and ground as described above.

Density Separation of Heavy Particulate Organic Matter

The heavy material which did not float at a density of 1.85 g cm^{-3} was backwashed onto a 20-μm nylon filter, and a vacuum was applied to remove excess water and polytungstate. The material retained on the filter was rinsed into 100-mL beakers with sodium polytungstate (2.22 g cm^{-3}) to a volume of 50 mL. The samples were allowed to separate overnight, after which we aspirated the heavy POM (hPOM) floating on the surface of the liquid. The sand fraction was recovered by backwashing the material that did not float at 2.22 g cm^{-3} onto a 20-μm nylon filter. The hPOM and sand fractions were rinsed with water, dried, and ground as described previously.

Carbon and Nitrogen Determination

Organic C and total N in the surface residue, coarse roots, whole soil, and SOM fractions were determined by dry combustion methods in a Carlo-Erba NA 1500 NCS elemental analyzer (Haake Buchler Instruments, Paterson, NJ). No carbonates were present in this soil. Carbon-14 concentrations in each fraction were measured by combusting subsamples in a Harvey Biological Oxidizer, model OX500 (R.J. Harvey Instrument Corp., Hillsdale, NJ). The ^{14}C released during oxidation was trapped in Harvey's ^{14}C cocktail and counted on the liquid scintillation analyzer. The ash content of each fraction was determined by loss of mass upon ignition in a muffle oven at 450°C for 20 h.

Uniformity of Carbon-14 Label Experiment

We conducted a separate incubation test to assess the uniformity of the ^{14}C label in the oat leaves and coarse roots. Labeled leaves and roots were collected, dried at 50°C, and then separately ground in a Wiley mill. We placed 25 g of pre-ashed silica in a 50 mL glass beaker and then stirred in 0.4 g of either the leaves or coarse roots. Three replicates of each treatment (oat leaves or coarse roots) were prepared. The material in each beaker received 1 mL of a general soil inoculum. Inoculum was obtained by shaking 250 g of soil in 500 mL of water for 20 min. The resulting suspension was centrifuged for 30 min and the opaque supernatant was collected as the inoculum source. In addition, each beaker received 1 mL of nutrient solution [1 g K_2PO_4, 0.25 g KCl, 0.25 g $MgSO_4$, and 23 mg $(NH_4)_2SO_4$ L^{-1}] and 1 mL L^{-1} of micronutrient solution (Wander et al., 1994). Additional water was added to bring the samples to 60% water-filled pore space based on the amount of water remaining after saturated sand freely drained for 24 h. Each beaker was placed in a sealable, 0.95 L jar along with a vial containing 8 mL of 0.5 *M* NaOH to capture CO_2–C as it was evolved. Controls were prepared by placing empty beakers and vials containing NaOH in three additional jars. We added 10 mL of water to the bottom of each jar to humidify the atmosphere within the jar and slow the rate at which the residue–silica mixtures dried out. The jars were sealed and placed in an incubator at 25°C. The traps were changed on Days 1, 3, 6, and after that weekly or semiweekly. Respired $^{14}CO_2$ was measured by adding Optima Gold scintillation cocktail to 1.0-mL aliquots taken from each trap and counting on the liquid scintillation analyzer. The total amount of respired CO_2–C (^{12}C + ^{14}C) was measured by titrating a 1-mL aliquot with 0.5 *M* HCl using a DTS800 Multi-Titration System (Radiometer, Copenhagen, Denmark). The specific activity (SA) of the CO_2–C evolved for each sample period was calculated by dividing the ^{14}C evolved by the total CO_2–C evolved. The results indicated the ^{14}C label was relatively uniformly distributed after the initial few weeks of the incubation for oat leaves and roots. From Day 0 to 30, the SA of the CO_2–C respired from leaf residue decreased and then remained constant through Day 210. The specific activity of CO_2–C from root residue increased from Day 0 to 15 and then remained constant through Day 210. These data show that, for the majority of the incubation time, both the leaves and roots were uniformly labeled.

Calculations and Statistical Analyses

On Day 0, we randomly selected three pots from the labeled surface residue treatment and measured the amount of ^{14}C in the surface residue ($D_{0\ residue}$), coarse roots ($D_{0\ roots}$), and whole soil ($D_{0\ soil}$). Similar measurements were made on three pots selected from the labeled (roots + soil) treatment. A baseline value for total ^{14}C in each treatment on Day 0 was calculated:

$$D_{0\ total} = D_{0\ residue} + D_{0\ roots} + D_{0\ soil}$$

$D_{0\ total}$ was 163 μBq kg^{-1} soil in the labeled surface residue treatment and 109 μBq kg^{-1} soil in the labeled (roots + soil) treatment. $D_{0\ residue}$, $D_{0\ roots}$, $D_{0\ soil}$, and $D_{0\ total}$ served as baseline measurements in the labeled surface residue and labeled (roots + soil) treatments to which subsequent sample dates were compared. On the remaining four sample dates, $D_{t\ total}$ was calculated for each of the treatments in a manner similar to that which we described above, except that cumulative $^{14}CO_2$ evolved was included in the total:

$$D_{t\ total} = D_{t\ residue} + D_{t\ roots} + D_{t\ soil} + D_{t\ CO_2}$$

Because the $D_{0\ total}$ values were different in the two treatments, comparisons between the treatments were made on a percentage basis.

The labeled surface residue treatment and the labeled (roots + soil) treatment were analyzed separately in this experiment. The pots were arranged in a completely randomized design. Three pots (replications) from each treatment were sampled on Days 0, 90, 180, 270, and 360. Regression models including linear and quadratic components were fit to characterize changes in each variable across time. Values for the LSD were calculated as described by Steel and Torrie (1997).

RESULTS AND DISCUSSION

Characteristics of Plant and Soil Materials

Characteristics of the plant and soil materials are given in Table 1. Carbon concentrations were relatively high and similar in surface residue and coarse roots, intermediate in large and small POM, and low in the hPOM, sand, and (silt + clay) fractions. The ash-free data show that the C concentration of leaves, roots, and POM fractions were about equal, indicating the observed differences in C concentration were primarily due to inequalities in the amount of inorganic material in each fraction. Nitrogen concentrations were higher in POM compared with the other plant and soil fractions. In contrast to our observations for C, differences in the N concentrations were accentuated by correcting the leaves, roots, and POM fractions for ash content.

Table 1. Characteristics of oat residue and soil fractions after 0 and 360 d of an incubation that simulated a no-till environment. Values on Day 360 followed by * changed significantly ($P < 0.05$) during the incubation according to linear regression analysis.

	C		N		C/N	
Compartment	Day 0	Day 360	Day 0	Day 360	Day 0	Day 360
	g kg^{-1} fraction					
Surface residue	385	208*	5.1	13.8*	75.8	15.2*
Coarse roots	387	359*	5.2	11.7*	74.7	30.8*
Large POM	310	302*	15.9	17.4*	19.5	17.5*
Small POM (<1.85)‡	260	280*	17.9	18.3	14.6	15.3*
Heavy POM (1.85–2.22)‡	13.7	14.0	1.0	1.0	13.7	13.8
Sand§	13.9	6.7*	1.2	0.6*	11.4	9.7
(Silt + clay)	13.7	14.8*	1.6	1.7	8.7	9.0
Whole soil	17.6	17.3	1.8	1.9	9.7	9.3*
	— Ash free g kg^{-1} fraction —					
Surface residue	471	487*	6.2	32.0*	75.8	15.2**
Coarse roots	467	473	6.3	15.4*	74.7	30.8*
Large POM	477	460	24.3	26.3*	19.5	17.5*
Small POM (<1.85)‡	468	508*	32.1	33.2	14.6	15.3*
Heavy POM (1.85–2.22)‡	447	473	32.4	33.7	13.8	14.1

† POM = particulate organic matter.
‡ Numbers in parentheses are density of SOM in g cm^{-3}.
§ Sand grains + organic material >2.22 g cm^{-3}.

Carbon-14 in Surface Residue, Roots, and Evolved Carbon Dioxide

To allow comparisons between the two treatments, we expressed the amount of ^{14}C in each compartment as a percentage of the total ^{14}C in the respective treatment on Day 0 (Tables 2 and 3). The recovery of ^{14}C after fractionation was generally >90% of the initial ^{14}C in each treatment (Tables 2 and 3). The amount of ^{14}C in the hPOM and sand fractions was relatively small and the two fractions were combined into one pool for discussion purposes.

The rate of ^{14}C loss from the surface residue and coarse roots was rapid at first and then slowed (Fig. 1). More than two-thirds of the ^{14}C was lost from the surface residue and coarse roots during the first 90 d of the incubation. At the end of the 360-d incubation, surface residue retained 11% of the original ^{14}C in residue ($D_{0\ residue}$) and coarse roots retained 7% of the original ^{14}C in coarse roots ($D_{0\ roots}$). The decomposition rates of surface residue and coarse roots were more rapid in our study compared with results from field experiments. Several investigators reported that for wheat straw, 14 to 57% of the surface residue mass is lost after 1 yr under field conditions (Brown and Dickey, 1970; Douglas et al., 1980; Holland and Coleman, 1987). We could identify no field studies that measured the decomposition rate of roots in no-till, but Buyanovsky and Wagner (1987) and Ghidey and Alberts (1993) reported that 65 to 78% of the root mass is lost after 1 yr in a disturbed soil.

The decomposition rates of the surface residue in our study were influenced by the decision to remove the stems and place only leaves on the soil surface. We made this choice for two reasons. First, the relatively high specific activity of the oat leaves would ensure a

Table 2. The distribution of surface residue-derived ^{14}C in various plant and soil compartments as a percentage of the total ^{14}C contained in the sum of all the compartments and d 0 ($D_{0\ total}$ = 163 μBq pot^{-1}).

	Day					Probability level†	
Compartment	0	90	180	270	360	Linear	Quadratic
	% of $D_{0\ total}$						
Surface residue	100	32.3	14.8	16.6	11.4	0.0001	0.0001
Coarse roots	0	0.2	0.1	0.1	0.1	0.7213	0.0036
Large POM‡	0	0.3	0.2	0.2	0.1	0.7654	0.0136
Small POM	0	0.9	1.9	1.7	3.0	0.0001	0.8246
(hPOM + sand)	0	0.5	0.7	0.8	1.0	0.0001	0.0760
(Silt + clay)	0	8.9	13.5	10.7	12.3	0.0001	0.0001
Total residue + soil§	0	43.1	31.2	30.1	27.9	0.0001	0.0001
Cumulative CO_2	–	51.3	61.1	65.0	66.2	0.0001	0.0001
	% Recovery¶						
	100	94.4	92.3	95.1	94.1		

† Level of significance (P) of linear and quadratic components in the regression model for each fraction.
‡ POM = particulate organic matter; hPOM = heavy particulate organic matter.
§ Sum of ^{14}C in surface residue, coarse root, and SOM fractions divided by the amount of ^{14}C contained in the surface residue and d 0.
¶ Sum of ^{14}C in all compartments divided by the amount of ^{14}C contained in the surface residue on d 0.

Table 3. The distribution of root-derived ^{14}C in various plant and soil compartments as a percentage of total ^{14}C contained in the sum of the compartments and d 0 ($D_{0\ total}$ = 109 μBq pot^{-1}).

	Day					Probability level†	
Compartment	0	90	180	270	360	Linear	Quadratic
	% of $D_{0\ total}$						
Surface residue	0	0.4	0.3	0.2	0.2	0.2798	0.2120
Coarse roots	61.6	10.3	5.6	5.1	4.2	0.0001	0.0001
Large POM‡	12.6	5.9	5.1	6.0	5.5	0.0001	0.0001
Small POM	3.2	5.0	5.6	5.8	6.5	0.0001	0.0681
(hPOM + sand)	1.7	1.8	1.9	2.5	1.6	0.4249	0.0389
(Silt + clay)	17.1	23.3	22.8	21.1	23.9	0.0109	0.0884
Total residue + soil§	96.2	46.7	41.3	40.7	41.9	0.0001	0.0001
Cumulative CO_2	–	39.3	46.9	53.7	56.1	0.0001	0.0001
	% Recovery¶						
	96.2	86.0	88.2	94.4	98.0		

† Level of significance (P) of linear and quadratic components in the regression model for each fraction.
‡ POM = particulate organic matter; hPOM = heavy particulate organic matter.
§ Sum of ^{14}C in surface residue, coarse root, and SOM fractions divided by the amount of ^{14}C contained in the (roots + soil) treatment on d 0.
¶ Sum of ^{14}C in all compartments divided by the amount of ^{14}C contained in the (roots + soil) on d 0.

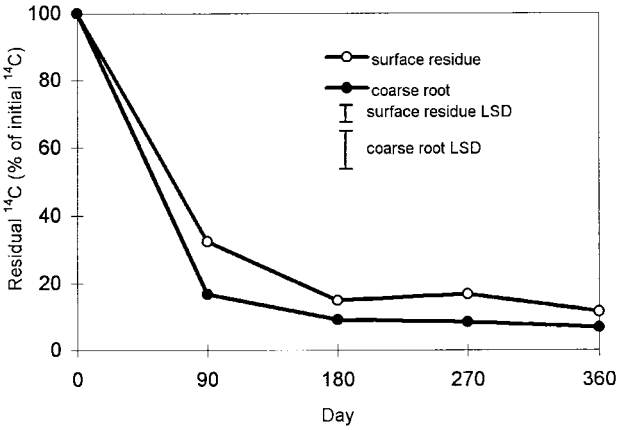

Fig. 1. Changes with time in the ^{14}C content of oat surface residue and coarse roots. Values are expressed as a percentage of the amount of ^{14}C contained in the surface residue or coarse roots on d 0 ($D_{0\ residue}$ = 163 μBq pot^{-1}; $D_{0\ roots}$ = 67 μBq pot^{-1}). Vertical bars indicate LSD at P = 0.05.

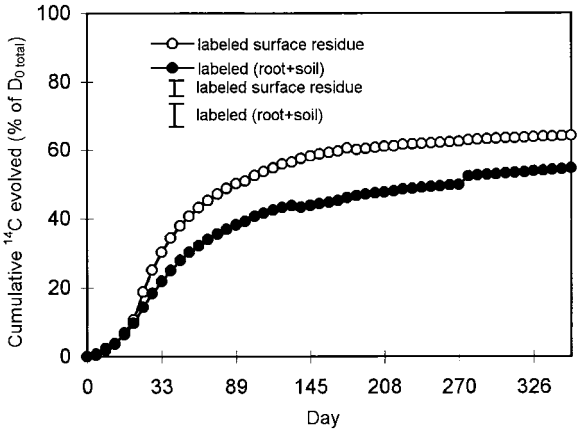

Fig. 2. Cumulative $^{14}CO_2$ evolved from pots containing either ^{14}C-labeled oat surface residue or ^{14}C-labeled (roots + soil). Values are expressed as a percentage of the total amount of ^{14}C in the respective treatments on Day 0 (Labeled surface residue treatment $D_{0\ total}$ = 163 μBq pot^{-1}; Labeled (roots + soil) treatment $D_{0\ total}$ = 109 μBq pot^{-1}). Vertical bars indicate LSD at P = 0.05.

strong, easily detectable signal in the SOM pools. Second, our objectives specifically addressed changes in soil C pools associated with biologically active soil processes, such as nutrient cycling and soil aggregation. Collins et al. (1990) reported that the half-life of wheat stems in soil is an order of magnitude greater than the half-life of wheat leaves. From this information, we calculated the potential maximum C input from stems to be nearly four times smaller than from leaves over a period of 1 yr. Therefore, we expected that the majority of the C entering biologically active soil pools during the year-long incubation would come from the leaves.

Cumulative $^{14}CO_2$ evolution is reported as a percentage of the $D_{0\ total}$ for the respective treatments (Fig. 2). Initially, the rate of $^{14}CO_2$ evolution was similar in both treatments. By Day 10, the rate of $^{14}CO_2$ evolution increased in the labeled surface residue treatment, but remained fairly constant in the labeled (roots + soil) treatment. Respiration rates began to slow in both treatments by Day 50 and were nearly constant after Day 130 in the labeled (roots + soil) treatment and after Day 150 in the labeled surface residue treatment. After 360 d, 66% of the initial ^{14}C contained in the labeled surface residue treatment and 56% of the initial ^{14}C in the labeled (roots + soil) on had been respired as CO_2.

Carbon-14 in Size and Density Fractions

A comparison of the two treatments indicates differences in the distribution of surface residue– and root-derived C among size and density fractions for all sample dates (Tables 2 and 3). The most notable difference was in the large POM fraction. It contained <0.3% of the initial ^{14}C ($D_{0\ total}$) in the labeled surface residue treatment compared to 5 to 13% of the initial ^{14}C in the labeled (roots + soil) treatment. More generally, large and small POM together contained 1 to 3% of the initial ^{14}C ($D_{0\ total}$) in the labeled surface residue treatment (Table 2). The (silt + clay) contained an additional 9 to 14% of the surface residue–derived C. In contrast, the large and small POM fractions contained 11 to 16% of the initial ^{14}C ($D_{0\ total}$) in the (roots + soil) treatment (Table 3). Addditionally, the (silt + clay) fraction from this treatment contained an additional 17 to 24% of the initial root-derived ^{14}C. The (hPOM + sand) fraction contained <3% of the ^{14}C in both treatments.

The ^{14}C content of many of the SOM fractions changed significantly during the incubation. In the labeled surface residue treatment, the amount of ^{14}C in the (silt + clay) fraction increased rapidly until Day 180 and then remained constant (Table 2). The (silt + clay) fraction includes not only decomposition resistant humic materials, but also more rapidly decomposable C pools including microbial biomass, some microbial metabolites, and soluble C (McKeague, 1971; Turchenek and Oades, 1979; Ahmed and Oades, 1984). Any ^{14}C additions to the (silt + clay) fraction after Day 180 may have been negated by losses from these labile components. In contrast, small POM–^{14}C in the labeled surface residue treatment increased linearly with time throughout the entire incubation period (Table 2). This accrual of surface residue–derived C in small POM suggests that the decomposition rate of this fraction is relatively slow even though it is a potentially rich source of C and N for microorganisms. The decomposition rate of small POM may be reduced because it is either physically or chemically protected from microbial attack.

In the labeled (roots + soil) treatment, large POM contained nearly 15% of the initial ^{14}C on Day 0 (Table 3). Large POM–^{14}C declined by 50% between Days 0 and 90, but small POM–^{14}C and (silt + clay)–^{14}C increased during the same time. This suggests that C is moving from the coarse root or large POM pools into the small POM and (silt + clay) fractions as decomposition proceeds. Changes in the ^{14}C content of large POM, small POM, and (silt + clay) associated material were minimal after Day 90 (Table 3). Losses from these pools due to decomposition may have been compensated for by additions from the coarse roots during this time, although most of the coarse roots were decomposed by Day 90. Alternatively, the decomposition rate of each of these pools may be relatively slow. We suggest that a portion of the root-derived C in soil is rapidly decomposed after plant senescence. Within a short time, the remaining root-derived C becomes stabilized either in chemically resistant forms or within aggregates that physically protect the C from microbial degradation.

SUMMARY AND CONCLUSIONS

After 360 d of decomposition, 66% of the ^{14}C contained in surface residue on Day 0 had been respired as $^{14}CO_2$, 11% of the original ^{14}C remained in residue on the soil surface, and 16% of the ^{14}C was in the soil (Table 2). Most of the surface residue–derived C in the soil on Day 360 was associated with the (silt + clay) fraction. Only a small amount of surface residue–derived C accrued in the POM fractions. In comparison, 56% of the initial ^{14}C in the labeled (roots + soil) treatment on Day 0 evolved as $^{14}CO_2$, and 42% of the ^{14}C remained in the soil after 360 d (Table 3). Most of the root-derived ^{14}C was associated with the (silt + clay)

and POM fractions. This supports the hypothesis that, in relatively undisturbed soil, POM is derived primarily from roots.

These data show clear differences in the partitioning of surface residue– and root-derived C during decomposition and imply that the beneficial effects of no-till on soil organic C accrual are primarily due to the increased retention of root-derived C in the soil. A large amount of surface residue–derived C was respired into the atmosphere as CO_2, while a greater proportion of root-derived C was conserved within the soil after 1 yr. The contribution of surface residue to SOM may be higher over the long term (i.e., >10 yr) due to C inputs from the relatively slowly decomposing stem tissue. However, we calculated the potential C input from stems to be nearly four times smaller than the amount contributed by leaves during our 1-yr incubation.

It should also be noted that the contribution of surface residue to SOM may be higher under field conditions where earthworms (*Lumbricus terrestris*) and other fauna feed on the surface residue and deposit casts throughout the soil. Furthermore, to limit the variables in our experiment, we minimized the movement of soluble C into the soil during the incubation by injecting water directly into the soil with a cannula rather than pouring water through the residue. As much as 10% of the C in wheat straw may be soluble (Collins et al., 1990) and, under normal field conditions, rainwater could leach this C into the soil and potentially increase the amount of surface residue–derived C in the soil.

These experiments were designed to examine changes in soil C pools during the initial period following the implementation of no-till. Early changes in the C pools are most likely to be manifested in biologically active processes, such as nutrient cycling and soil aggregation. Although our experimental conditions would not exist in situ under typical no-till conditions, the results are an important first step towards developing and testing hypotheses about the relative contribution of surface residue and roots to SOM in the field.

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